As agreed upon by the Trustees and BP, all samples collected for contaminant analysis during the sampling plan described below will be sent to Alpha Analytical or Columbia Analytical Services, unless they are designated to be archived. Samples for other analyses, if not archived, will be sent to the laboratories indicated in the plan below. ■

Each laboratory shall simultaneously deliver raw data, including all necessary metadata, generated as part of this work plan as a Laboratory Analytical Data Package (LADP) to the trustee Data Management Team (DMT), the Louisiana Oil Spill Coordinator's Office (LOSCO) on behalf of the State of Louisiana and to BP (or ENTRIX behalf of BP). The electronic data deliverable (EDD) spreadsheet with pre-validated analytical results, which is a component of the complete LADP, will also be delivered to the secure FTP drop box maintained by the trustees' Data Management Team (DMT). Any preliminary data distributed to the DMT shall also be distributed to LOSCO and to BP (or ENTRIX on behalf of BP). Thereafter, the DMT will validate and perform quality assurance/quality control (QA/QC) procedures on the LADP consistent with the authorized Quality Assurance Project Plan, after which time the validated/QA/QC-ed data shall be made available simultaneously to all trustees and BP (or ENTRIX on behalf of BP). Any questions raised on the validated/QA/QC results shall be handled per the procedures in the Quality Assurance Project Plan and the issue and results shall be distributed to all parties. In the interest of maintaining one consistent data set for use by all parties, only the validated/QA/QC-ed data set released by the DMT shall be considered the consensus data set. In order to assure reliability of the consensus data and full review by the parties, no party shall publish consensus data until 7 days after such data has been made available to the parties. Also, the LADP shall not be released by the DMT, LOSCO, BP or ENTRIX prior to validation/QA/QC absent a showing of critical operational need. Should any party show a critical operational need for data prior to validation/QA/QC, any released data will be clearly marked "preliminary/un-validated" and will be made available equally to all trustees and to BP (or ENTRIX on behalf of BP).

All materials associated with the collection or analysis of samples under these protocols or pursuant to any approved work plan, except those consumed as a consequence of the applicable sampling or analytical process, must be retained unless and until approval is given for their disposal in accordance with the retention requirements set forth in paragraph 14 of Pretrial Order # 1 (issued August 10, 2010) and any other applicable Court Orders governing tangible items that are or may be issued in MDL No. 2179 IN RE: Oil Spill by the Oil Rig "DEEPWATER HORIZON" (E.D. LA 2010). Such approval to dispose must be given in writing and

by a person authorized to direct such action on behalf of the state or federal agency whose employees or contractors are in possession or control of such materials.

This plan will be implemented consistent with existing trustee regulations and policies. All applicable state and federal permits must be obtained prior to conducting work.

Approval of this work plan is for the purposes of obtaining data for the Natural Resource Damage Assessment (NRDA). Parties each reserve its right to produce its own independent interpretation and analysis of any data collected pursuant to this work plan.

APPROVED:

FOR ROLAND GUIDRY

KD

11/02/11

Louisiana Trustee Representative: Date

Oct 14, 2011

BP Representative: Date

10/11/2011

NOAA Trustee Representative Date

(on behalf of all other trustees)

Mississippi Canyon 252 Spill
Oyster Sampling Plan
2011 Oyster Quadrat and Sediment Sampling

October 6, 2011

Introduction

A Technical Working Group ("oyster working group") of experts and trustee agency and BP representatives has been assembled to develop work plans appropriate to carry out both baseline (pre-injury) and post-impact assessments of oysters throughout the northern Gulf of Mexico. This document presents a follow-on phase and expansion of diver assisted oyster reef quadrat sampling and sediment sampling previously conducted as part of the Phase I Amendment 2 pre-assessment sampling plan in summer/fall 2010 for oysters in the north-central Gulf of Mexico.

Objective/Purpose

After reviewing observational data and limited analytical data generated from the field efforts implemented under the Phase I Pre-assessment work plan and its amendments and the Oyster Sampling Transition Plan (Transition Plan), continued injury assessment sampling is warranted to estimate the degree and spatial and temporal extent of potential injury to oyster abundance and biomass resulting from: 1) potential exposure of oysters to contaminants released into the environment as a result of the Deepwater Horizon Oil Spill; and/or 2) potential exposure of oysters to changes in water quality (e.g pH and salinity) resulting from actions undertaken by the state of Louisiana in response to the spill. In addition, these samples may help us further characterize potential impacts to oyster resources resulting from the opening of the Morganza and Bonnet Carre spillways by the Army Corps of Engineers in May 2011.

In summer 2010, the oyster working group collected quantitative samples from oyster reefs as part of its Phase I Amendment 2 plan to assess potential injury to oysters and oyster habitat resulting from the Deepwater Horizon oil spill and response activities conducted by the state of Louisiana to address the spill. Field teams used diver-assisted quadrat sampling methods to obtain those quantitative samples, which were then processed to provide estimates of oyster abundance and biomass by size class at a set of historically sampled locations across the Gulf of Mexico in Louisiana, Mississippi, Alabama, and Florida. The field effort also included collection of sediment samples for contaminant analysis at each site. A subsequent sampling plan, the Transition Plan, supplemented the Phase I locations with a randomly selected sample of sites in

Louisiana and Mississippi that expanded the geographic coverage of sampling within areas known or likely to contain oyster habitat, and expanded sampling in freshwater diversion areas in Louisiana (See Figure 1).[1] To date, however, no quadrat samples have been collected at the Transition Plan sites. The plan described in this document (hereafter, the "2011 Quadrat and Sediment Sampling Plan") involves resampling of the Phase I sites for oysters by quadrat and expands the sampling plan to collect quadrat samples from the Transition Plan sites in Louisiana and Mississippi as well. The plan also includes collecting sediment samples for contaminant analysis at all sites.

The results of the 2011 Quadrat and Sediment Sampling Plan will be used to support the assessment of any injury to oyster abundance and biomass and to inform and support restoration planning efforts.

The Plan specifically addresses the following topics:

1. Approach and rationale: This section describes the overall purpose and need for the 2011 Quadrat and Sediment Sampling Plan.

2. Health and safety: This section summarizes pertinent health and safety protocols applicable to this effort. It includes a number of procedures by reference, all of which should be carefully reviewed and adhered to by all team members.

3. Site selection: This section describes the proposed approach to identifying sites for evaluation.

4. Estimated Study Cost: This section provides an estimate of the cost of implementing the 2011 Quadrat and Sediment Sampling Plan.

Approach and Rationale

As noted previously in the Phase I plan, quadrat sampling provides valuable data on abundance and biomass of oysters and related fauna because it achieves a highly quantitative sample (i.e.

[1] Freshwater diversion areas (Figure 1) are areas under the influence of freshwater resulting from diversions of freshwater by Louisiana to meet salinity targets for fisheries and maintain vegetation health. Following the MC 252 spill, freshwater diversions were employed for an extended period of time in an attempt to keep oil away from the Louisiana coastline.

the area and effort of the sample are well defined).[2] Small dredges are the next best alternative and have been used in the Transition and Spring 2011 sampling plans. These plans employed a catch per unit effort (CPUE) dredging approach to rapidly and cost effectively obtain oyster abundance data at the randomly selected Transition Plan sites, data which can identify low or zero abundance areas.

Preliminary data obtained from the Phase I Amendment 2 quadrat sampling show low or zero abundance values generally across much of the Phase I study area, including areas that may have been impacted by contaminants from the Deepwater Horizon spill, and/or from freshwater diversions instituted by the state of Louisiana in 2010 in response to the spill. In addition, the preliminary results from dredge-based abundance assessments at Transition Plan sites show similarly low findings. These initial results, as well as potential impacts on oyster resources of freshwater from the Morganza and Bonnet Carre spillway openings in Spring 2011, warrant follow-up assessments of abundance and biomass metrics at the Phase I sites using quadrat sampling. Quadrat sampling at the Transition Plan sites would provide abundance and biomass per square meter values for these locations. This additional sampling will contribute to understanding of the geographical and temporal extent of injuries to the adult and juvenile oyster populations, and counts of spat-sized oysters from the quadrat samples will complement data on oyster settlement monitoring that the oyster working group has been collecting under the Phase I, Transition, and Spring 2011 plans.

Below is a summary of the key aspects of the 2011 Quadrat and Sediment Sampling Plan:

- The plan collects samples at locations previously mapped and sampled under the two prior DWH NRDA oyster plans. This includes Transition Plan locations in Louisiana and Mississippi that were characterized as known oyster habitat (i.e., they included either oyster reef mapped prior to the DWH spill or they were identified by State biologists to have a high probability of productive oyster habitat). It also includes Phase I sampling sites that were historically sampled prior to the DWH spill across all four states.

- The set of sample sites includes a non-randomly selected set of 81 historically sampled 200 meter x 200 meter grid cells from the Phase I plan located across Louisiana (40), Mississippi (19), Alabama (10), and Florida (12). It also includes up to 60 sites in both Louisiana and Mississippi from the randomly selected sample of the 600 meter x 600 meter grid cells (sites) from the Transition Plan. Finally, it includes a small set (up to 10 sites) of Transition Plan grid

[2] Quantitative sampling of oyster reefs is commonly accomplished by randomly placing on the reef a quadrat made of PVC that covers an area of a full meter square (1 m x 1 m), and then collecting all biota and other material encompassed within that quadrat up to a specified depth equivalent to about the size of one's hand.

cells in Louisiana characterized by higher frequencies of observed surface oiling in the months following the MC252 spill.

- Table 1 shows the metrics to be assessed and samples to be collected as part of the 2011 Quadrat and Sediment Sampling Plan. All samples will be taken in accordance with the protocols and standard operating procedures (SOPs) presented at the end of this document.

- Oyster abundance and biomass will be measured during one round of quadrat sampling. Quadrat sampling locations will be randomly selected from data on oyster reef locations from previous mapping efforts conducted under the Phase I or Transition Plans.

- Sediment samples at each site will be collected from locations randomly selected from soft bottom locations identified from data from previous mapping efforts conducted under the Phase I or Transition Plans.

- Material collected from the quadrats will be bagged and sent to Dauphin Island Sea Lab (DISL) for abundance counts and biomass of oysters and other observed fauna. Contaminant tissue samples for contaminant analysis that are collected from quadrats by DISL staff will be archived at DISL.

- In areas with scarce resources, quadrat samples may be supplemented by oysters collected via small dredges or tongs, in order to obtain sufficient oyster mass for a tissue contaminant sample. These oysters are not factored into the abundance and biomass measures.

Estimated samples from this activity

Field teams under this plan will sample up to 149 sites located across the northern Gulf of Mexico, where the term "site" refers to a sample grid cell as previously defined under either the Phase I or Transition Plans. Table 2 summarizes sampling activity in the 2011 Quadrat and Sediment Sampling Plan. Assuming adequate oyster resource exists and is accessible to the dive and sediment sampling teams, the target number of samples resulting from this activity are as follows:

- 596 oyster quadrats (4 per sample site) that will be analyzed for abundance and biomass measurements at Dauphin Island Sea Lab;
- 596 sediment samples (4 per samples site) that will be archived; and
- 596 oyster tissue samples (4 samples per site, 6 market-sized oysters or equivalent per sample) that will be archived.

- Priority for analysis of archived sediment and oyster tissue samples for contaminants will be determined under a separate analytical plan to be developed following review of chemistry results from samples collected during previous NRDA surveys.

Health and Safety

- The team leader and field crew parties should have completed all applicable health and safety training as directed by NOAA or state agency oil spill policy.
- All field team members must complete the NOAA safety training and documentation requirements as set forth in "Safety Requirements for All Personnel Working on NOAA-led NRDA teams for MS Canyon 252 Incident" (NOAA Safety Documentation Requirements.doc).
- All field team members should read all of the documents in the Safety directory on the case's NOAANRDA.org site.
    - Exception: if site collection activities do not include use of a boat or helicopter, then familiarity with the safety documents for these vehicles is not required.
- Each field team must submit a plan, not later than the night prior to going into the field. This plan must specify:
    - The team leader;
    - Names of all team members;
    - The sampling location(s)-- please use the grid coordinates as provided to your team by NOAA NRDA Field Ops staff or the NRDA Oyster Sample Location Coordinator;
    - What kind of sampling they are doing;
    - Expected arrival time at sampling area (daily);
    - Expected departure from sampling area (daily);
    - Team deployment date;
    - Team return date.
    - This information may be reported in one of two ways:
        - Fill out the Excel spreadsheet "Team Member Information Form – Sampling and Safety.xls" and send it to dwhnrdafieldops@gmail.com. Please use one tab for each team.
        - If you cannot submit this spreadsheet electronically, you can call in and report the information using this number: 1-504-656-6432
- Field teams must adhere to all procedures set forth in the most recent version of the MC252 Site Safety Plan ("NRDA_Ops_Safety Plan_08 04 11.docx").

- If participating in a cruise: Each cruise may have additional required health and safety procedures, which must be observed.
- Any encounters with protected species are to be reported to the appropriate authorities. Field crews are also to follow any guidance or BMPs provided by federal, states, or tribal historic preservation officers to avoid potential impacts to protected species or to historic or cultural resources. Any affected historic or cultural resources are to be reported to the appropriate authorities as described in such guidance or BMPs.
- Diving: SCUBA or surface-assisted diving, where used for sampling, will be conducted in accordance with the most recent version o f the Job Hazard Analysis for diving in the Field Ops Safety Plan, currently "Field Ops Safety Plan, Appendix A: Job Hazard Analyses." All divers will have SCUBA and scientific diver certification. BP/Cardno ENTRIX will not participate in SCUBA or surface-assisted diving but will assist with work on dive quadrat and sediment sampling boats.

Site Selection

No new sites were selected for sampling as part of the 2011 Quadrat and Sediment Sampling Plan; all sites to be sampled under this plan have been previously sampled under either the Phase I Plan or the Transition Plan. Sites included for quadrat sampling include all sites from both those plans, except for sites for which DWH NRDA mapping conducted at those sites found no substrate that could support oysters. Sites will be 200 m by 200 m grid cells (for sites from the Phase I Plan) in Louisiana, Mississippi, Alabama, and Florida or 600 m by 600 m grid cells (for sites from the Transition Plan) in Louisiana and Mississippi. The Phase I sites from summer 2010 included historic collection locations of the States' resource management agencies. Transition Plan sites were selected using a generalized random tessellation stratified (GRTS) sampling procedure. This plan will sample 149 sites overall, with 100 sites in Louisiana (40 from the Phase I Plan and 60 from the Transition Plan), 27 sites in Mississippi (19 from the Phase I Plan and 8 from the Transition Plan), 10 sites in Alabama (all from the Phase I Plan), and 12 sites in Florida (all from the Phase I Plan). Figures 1 through 5 show the locations of sampling for the 2011 Quadrat and Sediment Sampling Plan.

Sampling for the 2011 Quadrat and Sediment Sampling Plan consists of the following sampling events:

1. Quadrat Sampling: Quadrat samples will be collected at each site. Quadrats are made of PVC and are 1.0 m by 1.0 m (1 meter square).[3] The square meter quadrat may be sampled either by divers or by hand. Reefs in shallow subtidal areas (less than 0.5 m in depth) will be sampled by hand. Quadrats in deeper areas will be sampled using SCUBA or surface assisted divers. Quadrat samples will be collected at contact points randomly selected for each site where appropriate substrate (i.e., oyster reef) is present. Four spatially independent samples will be collected per cell and maintained as separate samples. Quadrat sampling will be used to determine oyster density, size frequency and biomass. Oyster tissue samples for contaminant analysis will be prepared from the quadrat samples and archived.

2. Sediment Sampling: Sediment samples will be collected at contact points randomly selected for each site where appropriate substrate (i.e., soft bottom) is present. Four spatially independent samples will be collected per cell and archived. See Appendix A, Section C for the detailed sediment sampling SOP, which is based on the findings of the draft letter report, "Draft Sediment Sampling Method Evaluation, NOAA NRDA Oyster Technical Work Group, Mississippi Canyon 252 Spill," dated January 12, 2011 and prepared by Stephen Emsbo-Mattingly of Newfields. The 2011 Oyster Quadrat and Sediment Plan includes a slightly revised SOP for sediment sampling, as compared to the Transition Plan. The SOP was revised based on field experience with push coring techniques gained through sediment sampling conducted under approved workplans in 2011 for other TWGs.

   The key difference involves use of core sampling as the initial sampling method under this plan. Under the Transition Plan, the SOP instructed the samplers conduct their first sample attempt using the modified van Veen grab sampler technique. Following a visual inspection and confirmation that each sample was of good quality and contained visible flocculent material, the samplers would proceed; however, if there was no visible flocculent or if the sample quality was poor, the samplers were instructed to collect a push core sample.[4] The 2011 Oyster Quadrat and Sediment Sampling Plan SOP reverses

[3] The Phase I Amendment 2 oyster sampling plans initially used ¼ meter square quadrats, and later employed 1 meter square quadrats at sites with very low resource to increase the likelihood of obtaining sufficient tissue for contaminant sample collection. The use of square meters at all sites will help streamline sampling and ensure adequate tissue is collected for contaminant analysis. Results from the two quadrat sizes are reasonably scalable with high correlation.

[4] Examples of poor quality samples include sediment rat holing, uneven sample surface, and water leakage.

this approach, indicating the push core sampler as the initial technique to capture samples, coupled with a visual inspection of the sample for flocculent and for indications of sample quality. If an appropriate and acceptable push core sample cannot be obtained after multiple attempts, the samplers are then instructed to try to obtain a sample with the modified van Veen grab sampler. The change in the order of collection methods should not affect the comparability of 2011 Quadrat and Sediment Plan sediment samples with previous sediment sampling events.

All samples for contaminant analysis will be archived. The prioritization of the analysis of samples will be determined pending a review of chemistry analyses of oyster tissue and sediment samples collected at or near these locations during previous NRDA surveys.

Cost Estimate

The estimated cost for this plan, excluding analytical costs for contaminant analysis of tissue and sediment analysis, is $1,400,098. The contaminant analytical costs will be estimated under a separate analysis plan for tissue and sediment samples. For details concerning cost estimates, refer to Table 3: Costs for 2011 Oyster Quadrat and Sediment Sampling Plan as well as the Excel file "Costs_2011_Quadrat_090811.xslx"

The Parties acknowledge that this budget is an estimate, and that actual costs may prove to be higher due to a number of potential factors. BP's commitment to fund the costs of this work includes any additional reasonable costs within the scope of this work plan that may arise because of any contingencies. The trustees will make a good faith effort to notify BP in advance of any such contingencies.

Table 1. Proposed Phase I metrics

Metric	Proposed Frequency of Sampling
Effect Metrics	
Oyster abundance (qualitative)	One event
Exposure metrics	
Tissue concentrations	One event
Sediment concentrations	One event
Oiling observations (qualitative)	Collected on each site visit

Table 2. Summary of oyster sampling procedures, maximum number of sites and replicates in the 2011 Quadrat and Sediment Sampling Plan.

Metric	Method	Potential # of sites				Max. Repl. per site	Est. Samples per event	Freq. of sampling	Total # of samples (estimate)
		LA	*MS*	*FL*	*AL*				
Adult and Juvenile Density	Quadrat	100	27	12	10	N = 4 quadrats	596	1	596
Tissue contaminant analysis	Oysters	100	27	12	10	N = 4 samples per grid cell	596	1	596
Sediment Contaminant analysis	Sediment	100	27	12	10	N = 4 samples per grid cell	596	1	596

Table 3. Costs for Oyster 2011 Quadrat and Sediment Sampling Plan

Item	Unit cost	Units	Units (teams * days or samples)	Total per event	# of events	Total cost (Event Total * frequency)	Detail
Quadrat Sampling				**$806,256**	**1.00**	**$806,256**	*Subtotal*
Divers				$471,833		$471,833	
Non-Diving Personnel				$198,667		$198,667	
Boat charges (4 boats)				$135,756		$135,756	
Quadrat Processing				**$220,520**	**1.00**	**$220,520**	*Subtotal*
Personnel				$214,560		$214,560	
Supplies				$2,980		$2,980	
Shipping and archive charges				$2,980		$2,980	
Sediment Sampling				**$337,402**	**1.00**	**$337,402**	*Subtotal*
Personnel				$198,667		$198,667	
Boat charges (4 boats)				$135,756		$135,756	
Supplies				$2,980		$2,980	
Sediment and Tissue Processing				**$11,920.00**	**1.00**	**$11,920.00**	*Subtotal*
Supplies	$5	Samples	596	$5,960.00		$5,960.00	
Shipping and archive charges	$5	Samples	596	$5,960.00		$5,960.00	
Cooler Rental (Maintained at Dauphin Island Sea Lab)	$2,000	month	6	**$12,000**	**2.00**	**$24,000**	*Subtotal*
Field/Lab Total						**$1,400,098**	
Total						**$1,400,098**	

Figure 1: 2011 Quadrat and Sediment Sampling Locations in Louisiana (CSA 1N, 1S, 3)

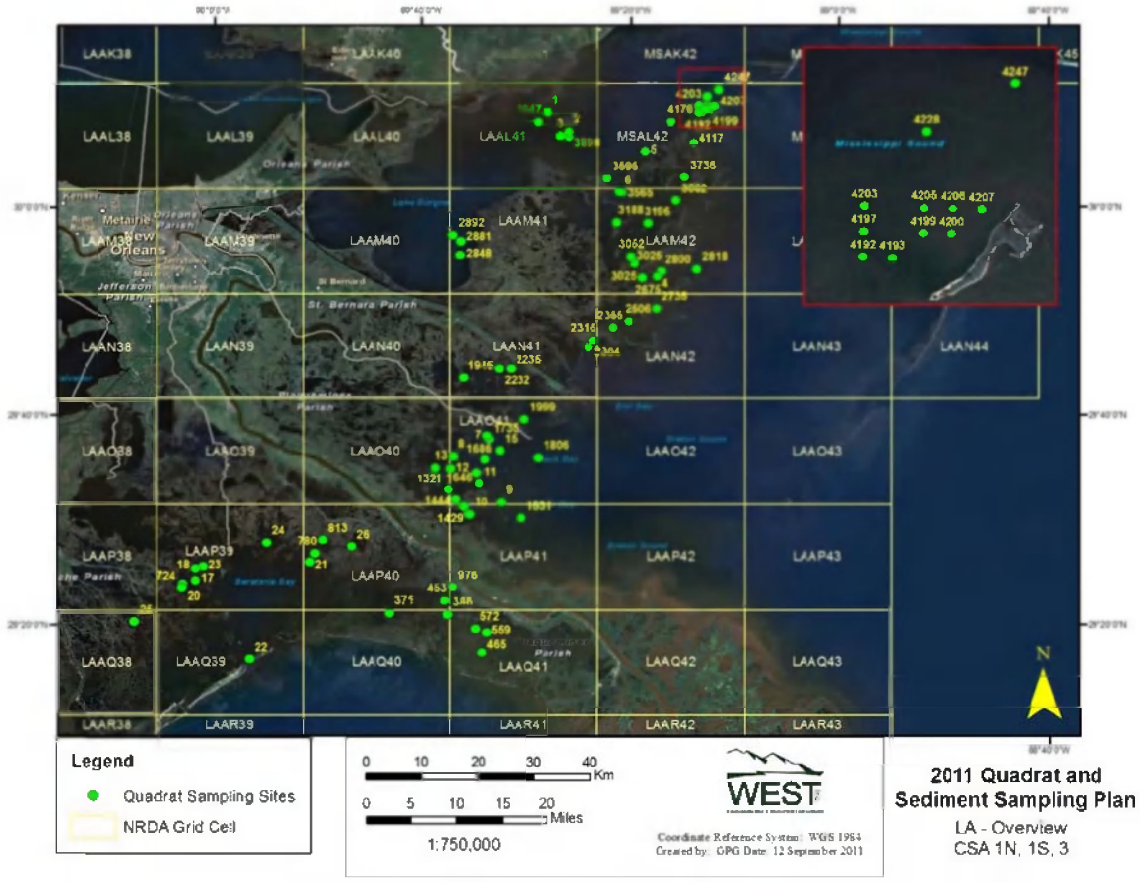


Figure 2: 2011 Quadrat and Sediment Sampling Locations in Louisiana (CSA 4 - 7)

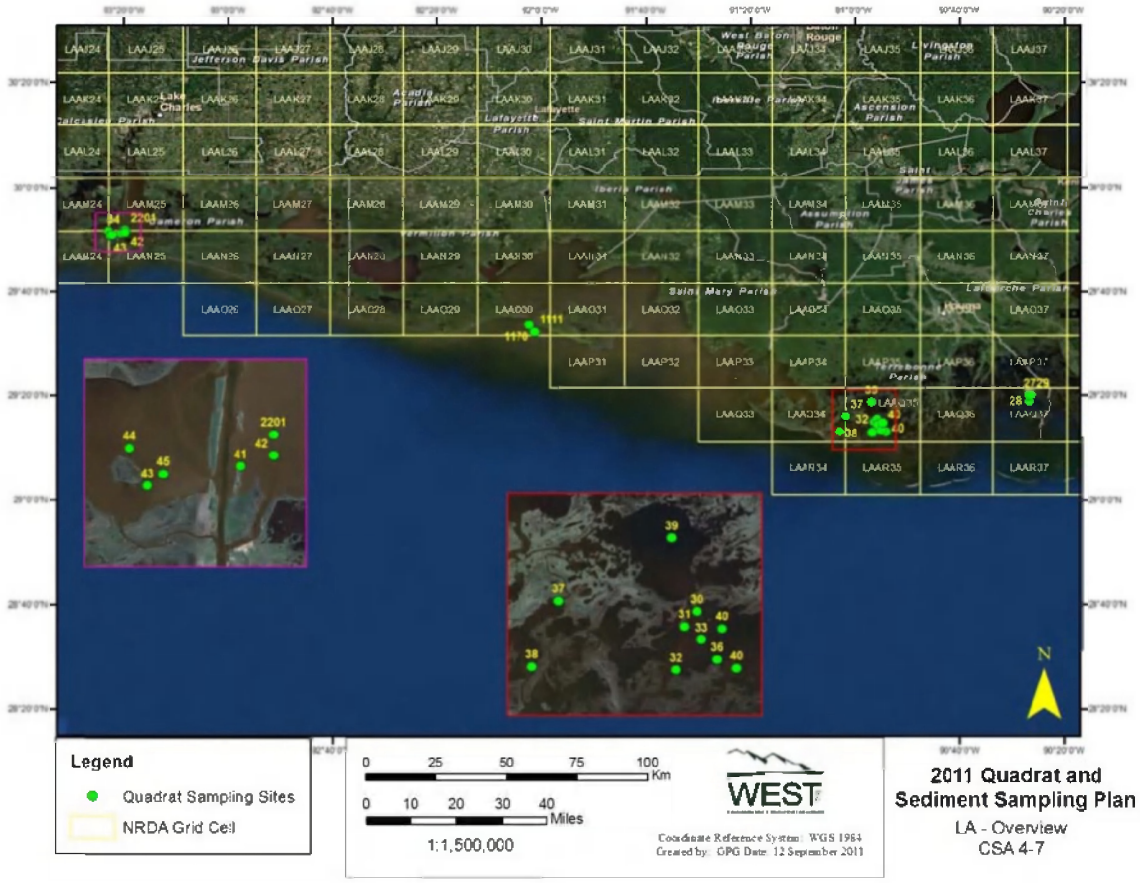


Figure 3: 2011 Quadrat and Sediment Sampling Locations in Florida

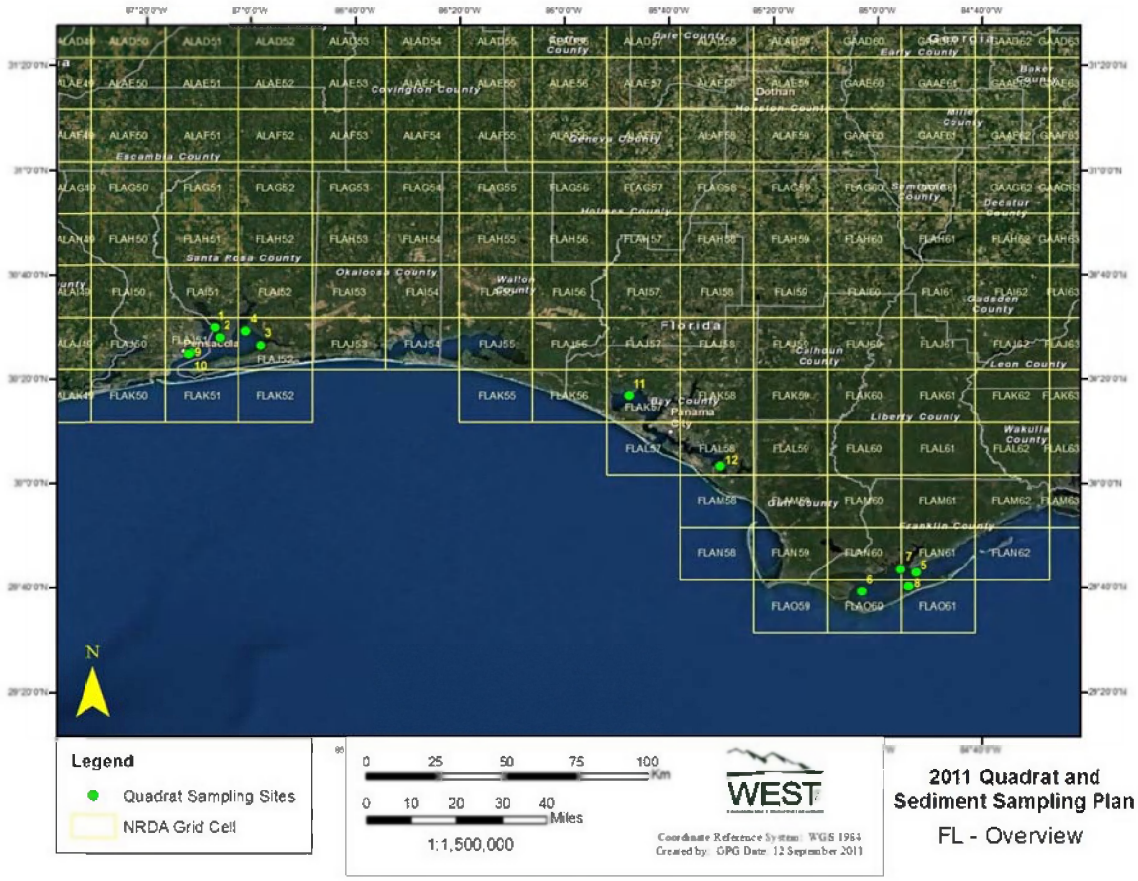


Figure 4: 2011 Quadrat and Sediment Sampling Locations in Mississippi

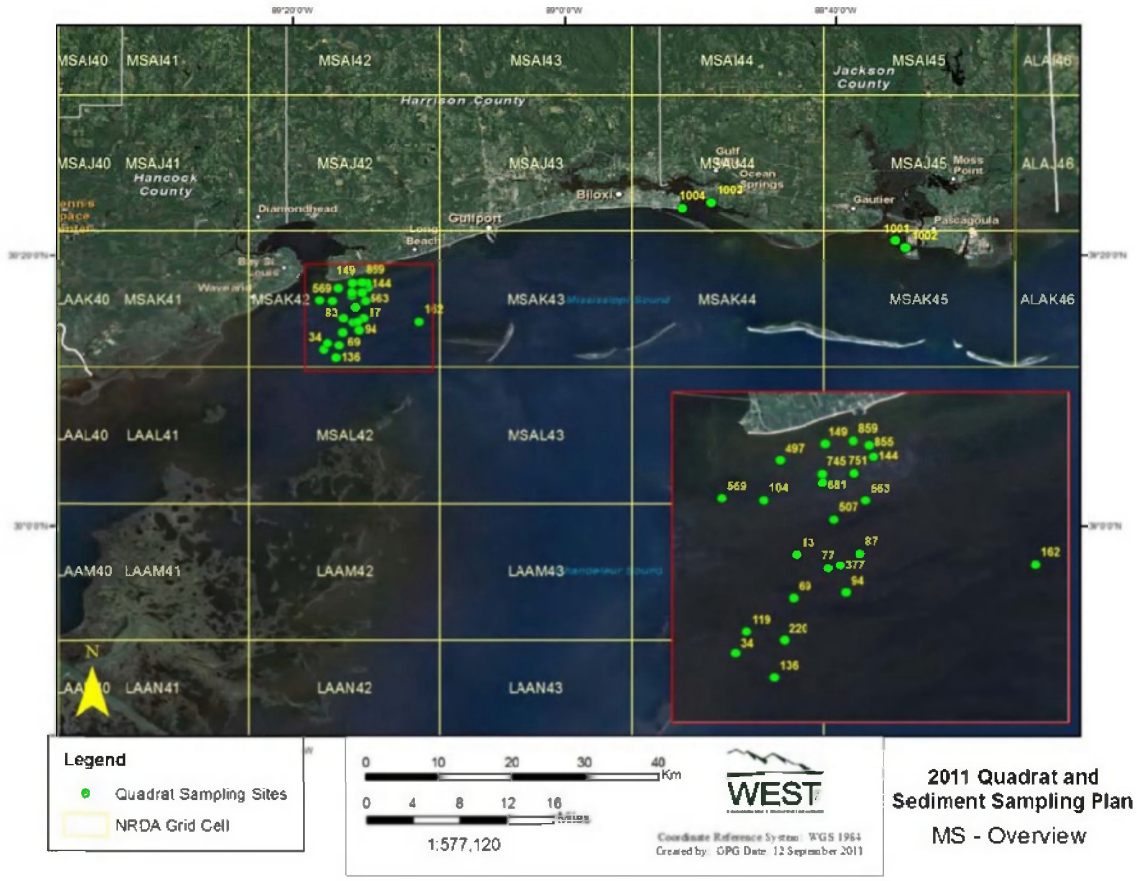
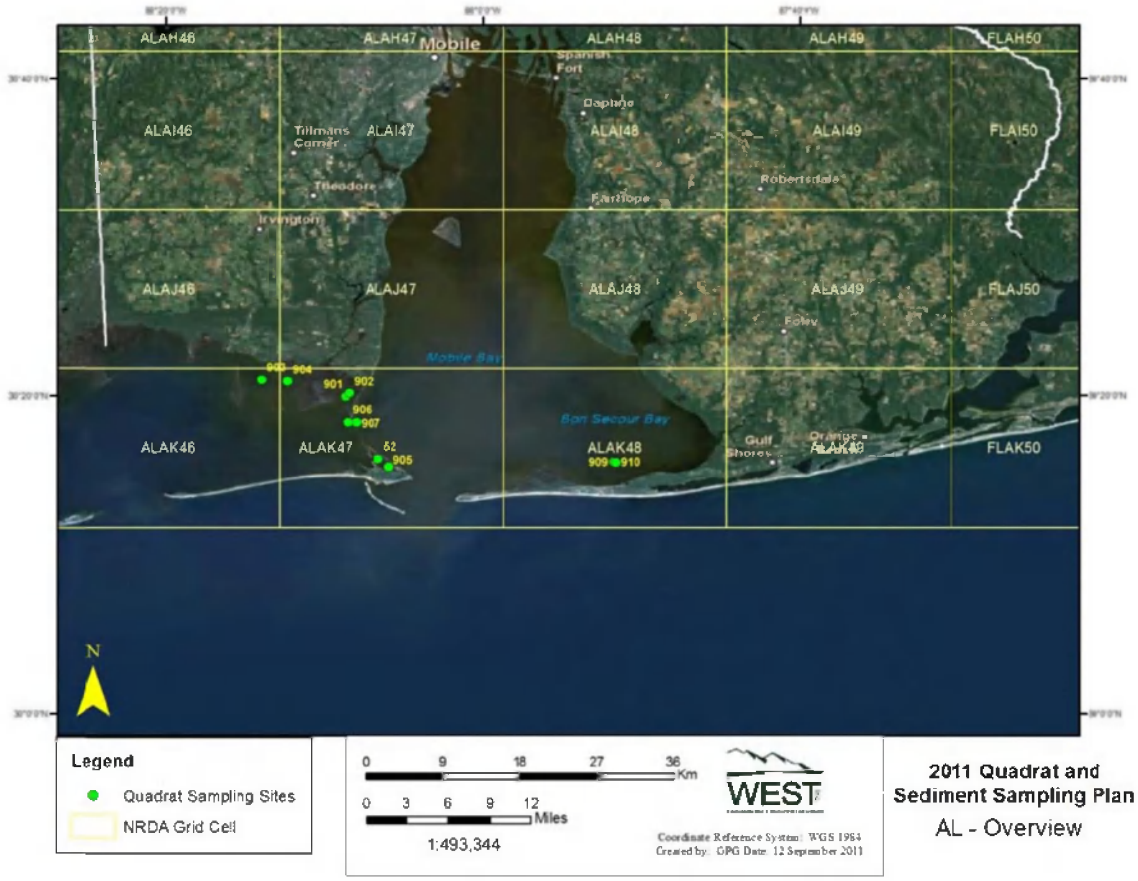


Figure 5: 2011 Quadrat and Sediment Sampling Locations in Alabama



Appendix A: Detailed Standard Operating Procedures (SOPs)
A. Juvenile and Adult Oysters (Settled Life Stages)

Field Sampling

Samplers should complete the **2011 Quadrat and Sediment Sample Plan – Site Visit Form,** the **2011 Quadrat and Sediment Sample Plan – Quadrat Form,** the **2011 Quadrat and Sediment Sample Plan – Site Summary/Signature Form**, and if needed, the **2011 Quadrat and Sediment Sample Plan – Dredge Form** or the **2011 Quadrat and Sediment Sample Plan – Tong Form** (see Quadrat_Field_Data_Version 1.1_09-26-2011.pdf). A unique sample ID should be given to each sample and prominently marked on the form according to the Oyster Sample Naming Convention (Appendix B). Sample codes should be recorded in the **2011 Quadrat and Sediment Sample Plan – Quadrat Form**, the **2011 Quadrat and Sediment Sample Plan – Dredge Form,** and/or the **2011 Quadrat and Sediment Sample Plan – Tong Form** datasheets and also in the **NRDA Sample Collection Form – Tissue/Wrack** (available on www.noaanrda.org).

1. *Site Description*

- Measure / Record:
    - Site name (**e.g., OYS-T-LA-A-465**)
    - Cell number **(GCID, e.g., 0020)**
    - Time of day and date.
    - Tidal depth (intertidal or subtidal)
        - If subtidal, estimate the depth in meters (record units) at the time of sampling.
        - Describe reef conditions – recent harvest, oiling, covered in mud, fouled, etc.

2. *Physical/Chemical Parameters*

- Measure and record:
    - Bottom and surface salinity
    - Bottom and surface water temperature
    - Bottom and surface dissolved oxygen (at subtidal sites only)
    - Ambient air temperature
    - Weather conditions
    - Oiled condition (None, Sheen, Scattered Deposits, Surface substantially covered, Surface completely covered or Deep Deposits).

After completing the above steps, move on to oyster sampling within the gridcell in accordance with the sampling methods below.

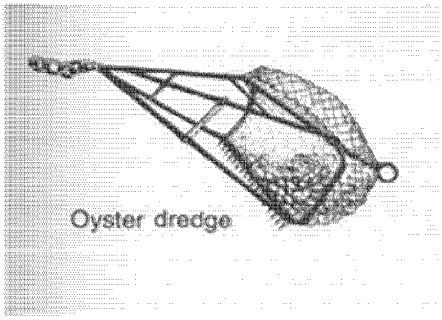
4. *Oyster Quadrat Sampling*

- Collect four individual quadrat samples per grid cell. Field teams will be provided with up to eight randomly generated contact points per grid cell. These contact points will be generated as a random sample of points from oyster reef transect segments identified in the mapping exercise.
- Determine coordinates via GPS. Ensure that you are within 5 meters of the contact point provided by NOAA NRDA Field Ops.
- Place 1 m^2 PVC quadrat frame directly at arm's length at a random spot (choose random side of boat to drop quadrat by pulling from a hat or spinning a compass) at the contact point. Do not favor abundant or sparse areas.
- Drop quadrat and have diver descend to depth (unless quadrat is in shallow water that does not require diving).
- Using tools when applicable, harvest all oysters 3-4 cm down into the reef (approximately the depth of a diver's gloved hand). You should not have to dig into the mud.[5]
- Place all material from the quadrat in burlap sack. When the quadrat collection is complete, secure sample to buoyed line.
- Divers are to signal to on-boat team that the quadrat has been collected. When divers are clear, on boat team pulls up the sample.
- Observe resource in the bag, and note the number of live oysters from the quadrat with respect to target collection goal. If necessary, dump collected material on tray to inspect.
- Return all material to burlap sack and close sack.
- Place the burlap sack in a plastic contractor bag.
- Samples should be tagged with an external (flagging tape with permanent marker) and internal tag (flagging tape with permanent marker) that prominently denotes sample code.
- The sample code should be constructed of the location ID, date, matrix, sample team leader code, grid cell id, and sample number along with information regarding sample type (for details, see the Oyster Sample ID Naming Convention, Appendix B).
- Hold animals on ice until delivered to intake team.
- Contact points should not be skipped. Retain all quadrat sample material and submit all quadrat samples to the intake lab.
- If the team fails to collect 6 oysters (>3") per cell or equivalent (after sampling 4 quadrats), then supplement the quadrat samples with dredging or tongs. Limit the period for additional sampling to 2 hours, and make sure to record the

[5] Collection of all material encompassed within the quadrat to a depth of approximately the size of a diver's gloved hand, as recommended in the SOP, will capture the preponderance of live and recently dead material present.

coordinates of dredge sampling on the dredge/tong field forms along with any notes. Bag only the live resource from the dredge/tongs for transmission to the lab and label accordingly.

- For dredge harvesting
    - Dredge harvesting using a 24 inch wide oyster hand dredge may be used to collect resource for contaminant or disease/gonad samples.
    - Deploy dredge from the beam of the vessel.
    - Drag dredge across the surface of the substrate for 3 minutes in a circular pattern around the contact point.
    - Record exact start and stop positions using a GPS.
    - Collect enough replicate dredge samples at sites chosen for quadrat sampling so that the required number of animals for analysis are obtained, if possible.
    - Place animals in a burlap sack.
    - Gently agitate the sack to remove excessive mud or debris.
    - Close sack.
    - Place the burlap sack in plastic bag.
    - Samples should be tagged with an external (flagging tape with permanent marker) and internal tag (flagging tape with permanent marker) that prominently denotes sample code.
    - The sample code should be constructed of the location ID, date, matrix, sample team leader code, and sample number along with information regarding sample type (for details, see the Oyster Sample ID Naming Convention, Appendix B).
    - Hold animals on ice until delivered to intake team.



- In areas where dredging is not possible because of logistical or permit difficulties, oyster tongs may be used to collect oysters.
    - Oyster tongs are generally 2-3 m long and constructed over two rakes welded or bolted together at the center point of the handles. The teeth on the rakes are generally 25 cm long and the head of the rake 1 m in length The rakes are juxtaposed to form a small basket when closed (local variations on oyster tongs are common and measurements need not be exact).

- Once at a site, the tongs can be deployed over the side of the boat. Once placed on the bottom the tongs are opened and closed repeatedly to dislodge oyster from a small area.
- After 6-10 opening and closing events, the tongs are used to collect the dislodged oysters into one grab. The tongs are held closed and the operator withdraws the handles from the water and places the contents on the deck.
- The entire procedure is repeated until the targeted number of oysters is collected.
- Four deployments should be made at each site; if no live oysters are collected, the boat should be repositioned 2 m away and the procedure repeated.

5. *Photographs*

See **NRDA Field Photography Guidance** (available onwww.noaanrda.org) for camera preparation and set-up prior to going into the field. –

- Photograph the operating GPS screen showing the date and time to synchronize the photos with the GPS track.
- Photograph site to describe oiling conditions.
- Collect a close-up photo of the reef showing individual oysters
- Photograph the entire reef.
- **DO NOT DELETE ANY PHOTOS**
- Document the pictures taken on the Oyster Reef Sampling Form
- Additionally, complete the NOAA NRDA Trustees Sampler Photo Logger form

6. *Collection and Disposition*

The individual who collected the sample should be noted on the field data form. If more than one person is involved, list the field party leader and the person who entered the data (if different). The final disposition of the sample should also be noted with an explanation of the amount of oysters retained for further analyses and the type of analyses (e.g., disease, histological analyses, contaminant). SOPs for these additional analyses are given below. Samples for tissue concentrations should also be collected and can consist of the same sample used to gather biological data (i.e., length frequency, etc.) if proper handling procedures (i.e. wrap oysters individually in aluminum foil, double bag, place on ice, etc.) are followed during and after sample processing.

Lab Processing

7. *Sample Processing: Abundance*

Samples will be brought to a non-field location for processing. Samples should be kept in a cooler with ice. Samples should be processed within 96 hours to ensure accurate characterization of live and dead oysters.

Regardless of sample method, both live and dead oysters should be enumerated by size category.

- Measure shell height (SH).
    - Using rulers (when oyster category is easily determined) and calipers (when fine scale separation of categories is needed to measure the distance from the umbo (small tapered end of the oyster) to the maximum limit of the shell.
    - Measure dead oysters in the same way.
        - Dead oysters are oysters that have no living tissue but are still in their articulated form (i.e., the shells are still hinged but no living oyster tissues is present also called "boxes"). These oysters will often appear opened or "gaped".
        - If the oyster is gaped and tissue appears to be undergoing decomposition, these oysters should be enumerated as a separate category
- Classify oysters by size:
    - spat (between 0.4 and 1 inch [10 - 25 mm] shell height),
    - seed or juvenile oysters (between 1 and 3 inches [25 – 75 mm]),
    - market size or "legal" oysters (> 3 inches [75 mm] shell height].

- Identify and enumerate associated biota.
    - Identifications and counts should be entered on separate lines under the "Other Species" category on the Oyster Reef Sample Form. Associated fauna should be archived with the quadrat sample. .

8. *Sample Processing: Biomass*

- Weigh living material:
    - Weighed in aggregate by size category.
    - Similarly, dead oysters should be weighed by category. Finally, associated species should be identified and weighed by taxon.
    - Dead oyster still with tissue will be classified as dead and weighed separately.

Equipment List

i. Random number table
ii. 2 PVC quadrats, dredge
iii. 3 sets of calipers and rulers
iv. 2 10-m long field measuring tapes (or laser range finder)
v. Spring scales (0-10g, 10-100g, 100 – 1000g, and 0-10 kg)
vi. Large 1 gallon Ziploc bags to separate subsamples for further analyses.
vii. Digital camera with extra batteries
viii. GPS with extra batteries
ix. Nitrile gloves (size M and L)
x. Small shovel / tool for separating oysters
xi. Waterproof data sheets (chain-of-custody, sample tracking, photo log, oyster reef sample form)
xii. Waterproof labels or tags
xiii. Waterproof pens
xiv. Flagging tape for external tags
xv. Burlap sacks for sample storage.
xvi. Plastic contractor-grade construction bags
xvii. YSI multimeter for DO, salinity

B. SOP for Tissue Collection for Contaminant Analyses (Based on Florida SERT Natural Resource Damage Assessment and NOAA Mussel watch protocols).

1. *Sampling Objectives*

(a) To document extent and duration of the area exposed to the spilled material. Bivalves uptake oil quickly, depurate them slowly, and can be used as "composite" samplers.
(b) To maintain the integrity the sample(s) during sampling, transport, and storage.

2. *Sample Size and pre-sampling activity*

(a) 20 g wet weight (composite of ~5 individual organisms).

(b) Clean dredges, knifes, etc. between samples. If no oil is visible wash in ambient water. If the equipment was obviously contaminated, rinse with Alconox solution. Collect rinsate for proper disposal.

(c) Take relevant photos at all sites before sampling.

3. *Sample Collection Methods*

(a) Collect primarily live animals (shells intact and tightly closed). Attached organisms are pried away from the substrate with a knife, trowel, etc. Infaunal samples should be rinsed with clean site water to remove sediment. Note the condition of dead animals if collected. If not collecting via quadrat sampling, animals can be collected by hand dredge or hand tongs. Once retrieved via the alternative method, animals should be handled in accordance with the below steps.

(b) The sampler handling the shellfish should wear nitrile or other non-contaminating gloves and change gloves after each sample to avoid cross-contamination. Record observations of any external evidence of contamination.

(c) Composite samples across all quadrats and/or dredges from a single site are recommended to provide enough sample weight to meet detection limit objectives and to average out the variations at a location among individual organisms.

(d) Individuals should be the same shell (or body) size. Record size range collected or save shells for later measurement. Same size is not as important if only for fingerprinting.

(e) Shellfish should not be opened in the field to minimize the risk of contamination. Rather, sets of whole organisms are collected together in burlap sacks.

(f) Place all individuals of the same species from a site in a certified-clean glass jar (without foil) or double Ziploc bags (with foil).

(g) For bags, the inner bag is labeled with marker pen and a waterproof sample label placed between the two bags. Jars are labeled on an adhesive label and directly on the lid. Use clear tape to protect the label.

(h) Avoid sources of contamination such as exhaust fumes and engine cooling systems on vessels. Work upwind of any exhausts. Segregate dirty/clean areas. Lay out clean substrates to work on and replace frequently. Take precautions so as not to introduce cross-contamination from oil on boots and shovels.

(i) If possible, sample least-oiled areas first, followed by the more contaminated areas to minimize risk of cross-contamination. Avoid sampling from creosoted pilings.

(j) Immediately place all samples in coolers on ice. Ship samples to the laboratory as soon as possible; samples should be received by the lab for processing or freezing within 7 days of collection. If holding samples for several days is unavoidable, samples may be stored frozen before shipping to the laboratory. Consult with dwhsampleintake@gmail.com for specific instructions; special shipping will be required to maintain samples in a frozen state until received by the lab.

(k) Use packing material around sample containers to prevent breakage during handling and shipping.

4. *Preservation/Holding Times*

Immediately place all samples in cooler and keep at 4°C. Freeze as soon as possible.

Please see the Analytical Quality Assurance Plan for the MS Canyon 252 (Deepwater Horizon) Natural Resource Damage Assessment (QAP) for further details on storage and holding times.

5. *Labeling, Documentation, and Other Considerations*

(a) On www.noaanrda.org, the NRDA Field Sampling Checklist generically summarizes pre- and post-field sampling tasks.

(b) Prepare sample labels as presented in NRDA Data Management Protocol for Field Sampling. If using jars, record the sample number on both the label and lid. IDs on sample labels must be complete and identical to IDs on the field sample form and the chain of custody. Jar labels

receive a protective layer of clear tape wrapped around the entire circumference of the container to secure the label and protect the writing.

(c) See the event-specific protocol documents for shipping to designated labs (NRDA Sample Shipping Instructions) and for chain of custody and sampling documentation instructions (NRDA Data Management Protocol for Field Sampling). Tissue sampling log sheets typically record sample number; date/time, location, GPS coordinates, species and tissue type.

(d) Documentation is critical; all field notebooks should be dated, signed, and preserved. If crossing out or correcting any entries, date and initial when making the changes. Original records will be gathered and archived.

(e) Record the presence of oil, weather conditions, etc. in field notes. Record GPS coordinates for each sample.

(f) Take relevant photographs of the sampling locations and sample collection itself if possible. Make sure each photograph or series can later be associated with the corresponding sampling location GPS (see NRDA Field Photography Guidance). Do not delete, open or alter any photos.

(g) All sampling, COC, shipping, GPS and photo files are uploaded to www.noaanrda.org.

(h) The labs have received instructions specifying sample processing and analytic methods.

6. *Analytical Methods*

The collected tissue samples should be analyzed in accordance with the MS Canyon 252 QAP. Specific suites of analytes to be measured include:

- Polynuclear Aromatic Hydrocarbons (PAH), including both standard and alkylated PAHs – see full list in Table 1.1a of the QAP, which also specifies the target method detection limits. If warranted, sterane/ triterpane biomarkers may also be quantified.

- Lipid content. Lipid content is defined as the percent of sample tissue extracted and remaining after solvent evaporation. It is used to normalize organic contaminants in tissues, to aid in spatial and temporal comparisons among samples.

Equipment List

i. Shovels and/or trowel
ii. Knife
iii. Dredges
iv. Tongs
v. Gloves (nitrile and knit Kevlar)

vi. Screen (for sieving out sediment)
vii. Aluminum foil
viii. Certified-clean glass jars
ix. Ziploc bags
x. Cooler and ice
xi. Marker pen
xii. Waterproof sample labels
xiii. Clear tape
xiv. Burlap sacks
xv. Coolers

PLEASE NOTE: Avoid sources of contamination such as exhaust fumes and engine cooling systems on vessels. Work up-wind of any exhausts. Segregate dirty/clean areas. Lay out clean substrates to work on and replace frequently.

C. SOP for Sediment Collection for Contaminant Analysis – Oyster Workplan

1. Objective

Collect a minimum of 240 mL of surface sediment (0-2 cm) including the flocculent material residing at the sediment water interface at predetermined locations. Collect a minimum of 240 mL of subsurface sediment (2-4 cm) for archive. Collect four individual sediment samples per grid cell. The first core will be analyzed for grain size and subsequent samples will be analyzed for contaminants. Field teams will be provided with up to eight randomly generated contact points per grid cell. These contact points will be generated as a random sample of points from soft bottom transect segments identified in the mapping exercise. Whenever possible, the sediment collection team will strive to capture and containerize a 375 ml sediment sample from each horizon in order to provide supplemental sample volume.

2. Field Equipment

Each crew should be deployed with multiple sampling devices to rapidly determine the general sediment type and apply the most appropriate sampling method to the field conditions. The basic sampling equipment should include the following elements.

Boat

The near-shore sediment sample collection methods will be deployed off small vessels (generally 12 to 40 ft fishing boats) with no winch, lifts, moon pools, or special anchors. However, these devices can be used, if available.

Weighted Tape Measure

The boats are typically equipped with an acoustic depth finder that generally has a precision of approximately +/- 1 ft. However, the depth finder may not be located where the sample is to be collected. It is recommended that the field team confirm the water depth with a precision closer to +/- 1 inch to help the sample operator gently lay the sampler on the sediment surface. The depth should be confirmed with an open reel tape measure with a weighted end (lead line) and saltwater-tolerant construction. For extremely fine sediments with a heavy flocculent layer, a small disc (approximately 6 inches in diameter) can be placed at the weighted end of the line to better identify the depth of the sediment-water interface. In waves or swells, the average water depth is estimated at half the distance between the top and bottom of a wave. Other comparable methods may be used as needed.

Samplers

The crews should be equipped with at least one grab sampler (e.g., van Veen) and one core sampler (e.g., push corer or piston corer). Diver corers may be used as a backup in the event that the first two options are not deemed successful. Table A-1 summarizes the features of the recommended samplers:

Table A-1. Specifications for Recommended Sampling Equipment.

Sampler	Dimensions	Dimensions	Surface Area	Maximum Surface Sediment Volume	Estimated Loss During Handling	Estimated Surface Sediment Volume per Attempt	Estimated Number of Attempts for 375 mL Sample
	In	cm	cm^2	cm^3 or mL	%	mL	
van Veen	8" x 6"	20.3 x 15.2	308	616	10	554	1
Ponar	6" x 6"	15.2 x 15.2	232	464	10	418	1
Ekman grab	6" x 6"	15.2 x 15.2	232	464	10	418	1
Piston or Push Core	3" diameter	7.62 diameter	45.6	91	0	91	4 to 5
Piston, Push or Diver Core	4" diameter	10.2 diameter	81.1	162	0	162	2 to 3

The sediment sampling teams will be trained and experienced with the various sediment sampling tools. The workplan investigation teams shall provide guidance to the sediment teams when evaluating the preferred sediment sampling methodology.

Modified Push Core or Piston Core Sampler

The push core sampler method allows the sediment sampling team the opportunity to visually inspect the sediment profile through the clear sample collection liner. The push core can be used to confirm the sediment structure observed in the van Veen sampler (e.g., how thick is the flocculent material at the sediment water interface) as well as to collect subsurface sediments. The push corer consists of a plastic core tube liner mounted on an aluminum head, and is fitted with extension rods that are sized to reach from the operator to the sediment surface. A one-way check valve or a ball valve works with suction to retain the sediment sample within the core liner. The push core system preserves the surface layers and simplifies the sampling procedure verses the piston core system. Alternately the piston core can create a greater vacuum and retain sediments within the core liner from deeper water depths.

For the piston core sampler, a piston slides within the core barrel, providing suction within the core tube and preventing sediment from flowing out the bottom of the sampler. The position of the piston is held at a fixed height above the sediment surface by a rope that extends to the operator. The use of the push core with the one-way flapper valve in place of the piston is the recommended sampler for this program.

The core liners are typically made of stiff clear plastic (e.g., polycarbonate). The volume of surface sediment is controlled by diameter of the core liner. The recommended piston core liner for silts and clays with organic matter is 10.16 cm diameter x 61 cm height (4" x 24") and the area sampled is 81 cm^2. The recommended piston core liner for flowable sands is 7.62 cm diameter x 61 cm height (3"x24") and the area sampled is 46 m^2. The liner thickness should be 0.16 cm (1/16"). The piston core head should be modified to accommodate either 3" or 4" core

liners in the field. Multiple cores are required for the production of large sample sizes. Three replicates of the 3" diameter core liner and two replicates of the 4" core liner should produce the minimum amount of sediment required for laboratory testing (240 mL), however the sample teams will be encouraged to obtain additional sediment volumes whenever possible.

To collect a sample, each push core tube is individually decontaminated as well as the core head. The push core tube is secured to the core head and lowered over the side using extension rods until the core tube is placed on the surface of the sediment. Hand applied force is applied to the extension rods and the top of the corer to drive the core tube into the sediment. It is recommended that a temporary mark (e.g., electrical tape) be placed on the extension rods indicating the water depth and targeted penetration depth. Once the desired depth is reached, the core tube and core head are pulled from the sediment and returned to the boat. At the boat the core tube should be capped below the water line with a clean plastic core cap, followed by recovering the core head, the core tube, the cap and the sediment profile are brought into the boat.

To collect a sample with the piston core, the core tube and core head is decontaminated as well as the piston plug and piston-tension rope. The tension rope is fixed to a stationary object, and force is applied to the top of the corer to drive it into the sediment. With a piston core the force may be manual or the core may be driven slowly into the sediments. It is recommended that a temporary mark (e.g., electrical tape) be placed on the extension pole indicating the water depth and targeted penetration depth. Once the desired depth is reached, the piston line is held tight against the extension rod and pulled up with the core. Care should be taken not to allow the piston line to move relative to the core tube. At the boat the core tube should be capped below the water line with a clean plastic core cap, followed by recovering the core head, the core tube, the cap and the sediment profile are brought into the boat.

van Veen Grab Sampler

For most substrate types, the van Veen sampler may be the preferred grab sampler. The van Veen grab sampler is a "clam-shell" device with opposing lever arms that close after the device contacts the sediment surface and the operator lifts the lever arms towards the boat. For hand deployment, a small van Veen grab is preferred, approximately 8" x 6" x 6" (20.3 cm x 15.2 cm x 15.2 cm). It is important to note the sampler dimensions to appropriately determine the area sampled. A grab that is 8" X 6" will sample an area of 308 cm^2. One grab should produce an adequate quantity of sample for laboratory testing and archive. It is recommended that the van Veen have screened doors on the top of the device to facilitate the movement of water through the device and reduce the bow wave that might otherwise push the flocculent material away from the sampler as it reaches the sediment water interface. In addition, the sampler should have a compound bucket profile that allows for the full area of the sampler to be sampled to the target depth. The device can include a stabilizing cage to assure perpendicular penetration. When needed, the device should accommodate the attachment of weights to facilitate the penetration of hard sediments. The location of these weights should not adversely affect the even closure of the clam-shell doors. The device should possess overlapping edges to retain water when closed. Finally, it is recommended that the rope on which the sampler is deployed to the sediment surface be marked (e.g., Sharpie or electrical tape) at every foot interval above the open mouth of the sampler so the sampling team can assure its gentle placement on the sediment water interface. The sampling personnel should handle the grab sampler carefully to avoid the accidental closure of the device on fingers, hands, or feet.

Ekman or Birge-Ekman

The Ekman grab sampler may be the preferred sampler for sediments with high amounts of flocculent material overlying the sediment surface. The van Veen sampler can work in these soft sediments, but care must be taken to avoid over-penetration. The Ekman sampler has spring-loaded jaws mounted on pivot points on the sides of a metal box. The dimensions are typically 6" X 6" X 6". The Ekman can be deployed by rope with a messenger, but the performance is improved by mounting it on a pole that allows the sampler to push the grab into the substrate. The jaws are activated from the surface by a push-knob at the top of the pole. Flaps on the top of the sampler open on descent to prevent a bow wave and close on ascent to reduce erosion or mixing. The pole may limit the water depths in which this sampler may be used. For the purposes of this survey, the Ekman should only be used at shallow sites with soft sediment. The sampling personnel should handle the grab sampler carefully to avoid the accidental closure of the device on fingers, hands, or feet.

Ponar Sampler

The Ponar grab sampler is a clam shell device with two opposing jaws with scissor arms that allow the two halves of the Ponar sampler to close after the sampler contacts and is subsequently lifted from the bottom. The Ponar sampler should be considered a backup device in the event that the van Veen sampler is not available, because the automated trigger mechanism is not as precise and accurate as the van Veen sampler. Ponar samplers are well suited for the collection of medium to moderately hard surface sediments. They are

manufactured in different sizes to address project specific objectives. The preferred dimensions are 6" X 6" X 6". Most Ponar samplers used to support the Oyster TWG programs are self-tripping, with a spring-loaded pin that releases when the sampler makes impact with the bottom. When closed, the jaws of the grab overlap preventing sediment loss. The top contains a screen that allows water to pass through the device and assure an even vertical decent and limits the creation of a bow wave as the sampler is lowered to the bottom. Rubber flaps cover the screens when the clam shell is closed and prevent sample loss when the device is lifted to the boat. The penetration of the sediment is largely controlled by the weight of the device and the force of the lever arms. Finally, it is recommended that the rope on which the sampler is deployed to the sediment surface be marked (e.g., Sharpie or electrical tape) at every foot interval so the sampling team can assure its gentle placement on the sediment water interface. The sampling personnel should handle the grab sampler carefully to avoid the accidental closure of the device on fingers, hands, or feet.

Diver Push Core Sampler

The Diver push corers are plastic tubes that SCUBA or surface assisted divers push into the sediment manually. They work best in soft to moderately hard sediment. The core liners are typically made of stiff, non-reactive plastic (e.g., polycarbonate). Once inserted into the sediment to a depth of approximately six inches, a polyethylene cap is placed on the top of the core. After a small hole is dug to the side of the corer, the diver places a second polyethylene cap on the bottom of the corer and returns to the boat with the core in its original vertical position. Alternatively, the diver may slide his/her hand under the core while it is extracted from the sediment. The volume of surface sediment is controlled by the diameter of the core. The recommended diver core liner is 10.16 cm diameter x 61 cm height (4" x 24") and the area sampled is 81 cm^2. The liner thickness should be 0.16 cm (1/16"). Multiple cores are required for the production of large sample sizes. Two replicates of the 4" core liner should produce the amount of sediment required for laboratory testing (a minimum of 240 mL). At every sampling location, the diver should descend with a basket capable of holding the cores in an upright orientation. The basket should contain 4 core liners and 8 core caps. One of these core liners is a backup in the event that the diver suspects problems with one of the initial 3 replicate cores.

3. Tiered Approach for Sample Collection

The field team will use a tiered approach for determining the most appropriate method for sample collection. For most substrate types the push core sampler should be the first sampler attempted for work associated with the Oyster investigation workplans. Upon recovery, the operator will determine if the sample is acceptable. If an acceptable sample cannot be collected, either the Ekman (in very soft sediment) or a van Veen grab sample should be attempted.

Acceptable Sample Criteria

An acceptable sediment sample consists of at least 8 cm in a core sampler or 5 cm in a grab sampler (>2") with an intact surface layer. Acceptable samples should not have sediment in direct contact with the doors of the grab samplers and should not have sediment pushing

through the door screens. If the overlying water drains out of the sampler, it should remain clear and should not significantly erode channels in the recovered sediment. A flocculent layer should appear on the surface when one is known to exist.

A sediment sample should be considered unacceptable if the surface layer contacted the top of the sampler or exhibited obvious features of mixing (e.g., no difference between the surface and subsurface when differences were known to exist). The absence of flocculent material in the grab samples should be confirmed by collecting a core sample. The van Veen sample should be recollected if the core sample exhibits significantly more flocculent material than the van Veen sample. An acceptable quantity of sediment should be collected within approximately 10 attempts. If this is not possible, the samplers should assess the samples to determine the reasons for poor performance and consider either an alternative location or alternative sampler.

3.1 Sediment Collection with the Push Core Sampler:

The push core will be the initial sample technique for the Oyster workplan. A one-way valve is installed on the air-water relief line from the core head, to create a vacuum during core removal. The simplicity of the one-way valve improves the ease of use and efficiency of the sampler. The valve is attached to the core head in a vertical position allowing air and water to exit as the core tube is pushed into the sediment and providing suction to hold the sediment in place as the tube is pulled upwards. Hard objects such as rocks, oysters, or shell-hash may impede penetration to target depth. Using a larger diameter tube may help avoid the interference of hard objects. Multiple attempts needed to obtain sufficient sediment quantities for analysis should be made, moving up-current from previous attempts, until sufficient volume is collected or it is determined that push-core sampling is not feasible in the area. A minimum of two core samples and preferably four core samples should be collected if using a 4"-diameter core tube; a total of 8 core samples will be needed if using a 3"-diameter core tube. Core samples should be collected with 30-cm to 60-cm core tubes and should be pushed into the sediment a minimum of 15-cm, or to the point of resistance (when the core cannot be pushed any further). A hammer (e.g., mallet or slide hammer) can be used to drive the core further into sediment, however, care should be taken not to drive the core in to the point that it cannot be pulled out by hand. Also physical agitation by hammer may disturb the core profile and should be minimized.

It may be necessary to conduct diver-operated coring in these situations as sample location can be chosen to avoid hard objects.

The following table (Table A-2) can be used to determine when to use a 3" diameter versus 4" diameter core tube for sampling. Alternative configurations can be used if comparable or better sample integrity can be achieved.

Table A-2. Piston Core Configuration.

Sediment Type	Texture	Diameter (inches)	Target Length (inches)
Organic Silt	Soft	4	12 - 18
Silty Sand	Intermediate	4	18
Sand	Hard	3	12-18
Shell or Veg	Hard	3 or 4	12-24

The procedure for collecting a sample using the push corer follows:

1. Upon arrival at the station, the field team should begin by confirming the water depth with a weighted tape measure and record the depth along with station coordinates. The type of sediment felt when measuring depth should be noted.
2. It is important to collect a core sample from a stable platform. In instances of strong wind or current the boat should be anchored to maintain location and for ease of sampling. The core tube must remain as close to vertical as possible to collect an acceptable sample.
3. Decontaminate core tubes and caps using a three-step field decontamination process. Ensure that all surfaces are free of visible sediment. Thoroughly wash all surfaces with soapy water and a bristle brush and then rinse with deionized water. Follow the DI water rinse, continue with a rinse on all surfaces with site water. If site waters are not considered suitable for a site water rinse, deionized water may be used solely.

   Alternatively, core tubes may be pre-cleaned by the analytical laboratory and wrapped in aluminum foil. In such cases individual core tube should be used to collect each sediment sample at a given station.
4. The push core head is a cylindrical piece of aluminum, which has a threaded hole at one end to receive the push rods. The first push rod should remain attached to the piston head. Additional rods may be added to accommodate the water depth at the station. It is recommended to mark the rods with one-foot increments.
5. Attach the core tube to the head using hose clamps and add push rods as needed to the other end of the head. Lower the push core sampler into the water allowing the core tube to fill with water. Air and water should come out of the one-way valve on the side of the piston head.
6. Gently lower the core tube until it contacts the sediment surface, being careful not to disturb the flocculent layer and keeping the tube and rods vertical. Note the location of the waters surface on the side of the extension rod and note the mark 15-cm (depending on the desired length of core) above the water surface. Push the sampler

straight down using hand force if possible or, if necessary, a small sledge or slide hammer, until the second mark is reached.

7. Gently pull the sampler straight up removing push rods as needed. Place a cap on the bottom of the tube when it reaches the surface of the water. The tube can then be set on the boat and the hose clamps and head can be removed. In softer sediments, the cap will need to be placed over the bottom quickly to avoid the loss of sediment from the bottom. Some loss is acceptable if the top 5 cm (2") are primarily undisturbed.
8. Rinse the outside of the core tube with water, then measure and photograph the core.
9. If the sample is considered acceptable using the criteria listed above, remove the piston core head by releasing the hose clamps. Remove the overlying water using a siphon or other similar means, taking care not to remove the flocculent layer.
10. Using a clean stainless steel spoon, collect the top 2 cm of the sample. Depending on the length of the tube and the sediment core it may not be possible to reach the sediment surface with a spoon. If the sediment is out of reach, the core tube can be cut with a hack saw one inch above the sediment surface, or the sediment can be pushed up from the bottom using an extruder until the surface sediment is reachable with a spoon. If this core is the first sample, label it as the grain size sample. If this core is the second acceptable sample, label it as the shallow contaminant sample.
11. Repeat the previous procedure for the collection of the next deeper sediment layer (2-4 cm) for archive. Label this sample as the deep contaminant sample.
12. Process samples in a manner consistent with the instructions in Section 4.
13. If the sample is not acceptable, remove the tube from the head, rinse clean of sediment away from sampling site and redeploy.
14. If the first sample is acceptable, it will be the grain size sample and subsequent samples will be contaminant samples. If the first sample is not acceptable, take two more core samples. If these samples are acceptable, the second sample should be labeled for contaminant analysis and the third sample should be labeled for grain size analysis. If, after three attempts, all samples are unacceptable, follow the protocol below for grab samples.

3.2 Sample Collection with the van Veen Grab Sampler:

1. Upon arrival at the station, the field team should begin by confirming the water depth with a weighted tape measure and record station coordinates.
2. Decontaminate the van Veen grab sampler using a three-step field decontamination process. Ensure that all surfaces are free of visible sediment. Thoroughly wash all surfaces with soapy water and a bristle brush and then rinse with deionized water

followed by site water. If site waters are not considered suitable for a site water rinse, deionized water may be used solely for the rinse process.

3. Ready the sampler for deployment. Ensure that the screened doors are locked shut. Set the sampler by lifting up on the chain at the shackle. Clip the ring in the pelican hook. As the grab is lifted, the jaws of the grab will open.
4. Lower the grab steadily to the sediment surface. It is important to lower the sampler slowly to the bottom, at a rate no greater than 1 ft/sec to prevent a bow wave. If the site is in deeper waters, the sampler may be lowered more rapidly until approximately 10 ft. above the bottom. The calibrated 1-foot marks on the rope will help the sampling personnel assure a gentle placement.
5. To collect a sample, allow the line to go slack once the sampler is on the bottom. Then, pull up slowly on the line, lifting the grab sampler causing the buckets to close. In firm substrate types, it is good to allow the sampler to "rest" on the bottom for approximately 30 seconds to allow it to penetrate the bottom. In softer substrate, minimize the contact time to prevent over-penetration.
6. As the sampler is retrieved to the sampling vessel, take care not to accidentally open the grab. The sampler can be hoisted by the chains or by one of the arms. Once retrieved, open the screened doors to inspect the sample.
7. If the sample is considered acceptable using the criteria listed above, remove the overlying water using a siphon or other similar means, taking care not to remove the flocculent layer. The unused overlying water can be returned to the sampling area. Using a clean stainless steel spoon, collect the top 2 cm of sample taking care not to collect sediment in direct contact with the sampler walls.
8. Using a clean stainless steel spoon, collect the next deeper sediment layer (2 - 4 cm) for archive. Take care not to collect sediment in direct contact with the sampler walls. Take precautions, like removing extra water or sediment to minimize the amount of sediment from the 0-2 cm interval that mixes with the 2 - 4 cm interval.
9. Process samples in a manner consistent with the instructions in Section 4.
10. If the sample is not acceptable, open the jaws of the grab, rinse clean of sediment and redeploy. If the sample has washed out, check the jaws for any debris preventing closure.

3.3 Sediment Collection with the Birge-Ekman Grab Sampler:

1. Upon arrival at the station, the field team should begin by confirming the water depth with a weighted tape measure and record station coordinates. Ensure that the water depths are within the capabilities for the Ekman sampler (generally ≤4 ft. with a standard pole).
2. Decontaminate the Ekman grab sampler using a three-step field decontamination process. Ensure that all surfaces are free of visible sediment. Thoroughly wash all

surfaces with soapy water and a bristle brush and then rinse with deionized water. Follow the DI water rinse with a rinse on all surfaces with site water. If site waters are not considered suitable for a site water rinse, deionized water may be substituted as the sole rinse step.

3. Ready the sampler for deployment. Set the sampler by pulling the jaws open and place the cable loops over the small pin near the triggering mechanism. Take care to keep fingers out of the closing path of the jaws.
4. Lower the grab to the sediment surface. Based on the lead line measurement, use depth marks on the sampler pole to determine when the sampler is at the sediment surface. In firm sediments, the sampler should push the grab into the sediment to a depth of approximately 4 to 5 inches. The Ekman grab is 6 inches in depth and care should be taken to avoid over-penetration. In soft sediment, little force will be required to push the sampler to the desired depth. In very fine sediment with an extensive flocculent layer, there may be little to no resistance as the grab is lowered into the substrate. In such cases, it is important to use the lead-line measurement to determine the maximum depth to lower the grab.
5. Once the grab sampler is in position, push the trigger at the top of the pole.
6. Retrieve the sample by pulling the pole slowly and steadily to the surface. Care should be taken keep the sample upright and keep the pole in the vertical position. As the sampler is retrieved to the sampling vessel, take care not to accidentally open the grab. Once retrieved, open the doors to inspect the sample.
7. If the sample is considered acceptable using the criteria listed above, remove the overlying water using a siphon or other similar means, taking care not to remove the flocculent layer. Using a clean stainless steel spoon, collect the top 2 cm of sample taking care not to collect sediment in direct contact with the sampler walls.
8. Using a clean stainless steel spoon, collect the next deeper sediment layer (2 - 4 cm) for archive. Take care not to collect sediment in direct contact with the sampler walls. Take precautions, like removing extra water or sediment to minimize the amount of sediment from the 0-2 cm interval that mixes with the 2 - 4 cm interval.
9. Process samples in a manner consistent with the instructions in Section 4.
10. If the sample is not acceptable, open the jaws of the grab, rinse clean of sediment and redeploy. If the sample has washed out, check the jaws for any debris preventing closure.

3.4 Sediment Collection with the Ponar Grab Sampler:

1. Upon arrival at the station, the field team should begin by confirming the water depth with a weighted tape measure and record the depth along with station coordinates. In instances of strong wind or current, the boat should be anchored to maintain location and for ease of sampling.

2. Decontaminate the Ponar grab sampler using a three-step field decontamination process. Lock the grab in the open position with the safety pin. Ensure that all surfaces are free of visible sediment. Thoroughly wash all surfaces with soapy water and a bristle brush and then rinse three times with site water. Follow the site water rinse with a rinse on all surfaces with deionized water. If site waters are not considered suitable for a site water rinse, deionized water may be substituted.
3. Ready the sampler for deployment. Ensure that the screened doors are locked shut. Set the sampler by replacing the safety pin with the spring-loaded pin, and applying vertical tension on the arms of the grab to keep the pin in place.
4. Lower the grab steadily to the sediment surface. It is important to lower the sampler slowly to the bottom, at a rate no greater than 1 ft/sec to prevent a bow wave. If the site is in deeper waters, the sampler may be lowered more rapidly until approximately 10 ft. above the bottom. Uneven lowering or sudden changes in tension on the line can cause the spring-loaded pin to prematurely fire. The calibrated 1-foot marks on the rope will help the sampling personnel assure a gentle placement.
5. To collect a sample, allow the line to go slack once the sampler is on the bottom. It may be necessary to gently shake the line, moving the arms of the grab slightly and reducing the pressure on the pin causing it to trigger. Then pull up slowly on the line, lifting the grab sampler and causing the buckets to close. In firm substrate types, it is good to allow the sampler to "rest" on the bottom for approximately 30 seconds to allow it to penetrate the bottom. In softer substrate, minimize the contact time to prevent over-penetration.
6. As the sampler is retrieved to the sampling vessel, take care not to accidentally open the grab. The sampler can be hoisted by the line or by squeezing the arms together above the hinge on the arms. Once retrieved, open the screened doors to inspect the sample.
7. If the sample is considered acceptable using the criteria listed above, remove the overlying water using a siphon or other similar means, taking care not to remove the flocculent layer. Using a clean stainless steel spoon, collect the top 2 cm of sample taking care not to collect sediment in direct contact with the sampler walls.
8. Using a clean stainless steel spoon, collect the next deeper sediment (2 - 4 cm) for archive. Take care not to collect sediment in direct contact with the sampler walls. Take precautions, like removing extra water or sediment to minimize the amount of sediment from the 0-2 cm interval that mixes with the 2 - 4 cm interval.
9. Process samples in a manner consistent with the instructions in Section 4.
10. If the sample is not acceptable, open the jaws of the grab, rinse clean of sediment away from sampling site and redeploy. If the sample has washed out, check the jaws for any debris preventing closure.

3.5 Sediment Collection with the Piston-Core Sampler:

The piston core should be used if the push core, van Veen or other grab samplers are not able to recover an acceptable sample after about ten attempts. A one-way valve is recommended in place of the rubber piston normally used. This will improve the ease of use and efficiency of the sampler. The valve is attached to the core head in a vertical position allowing air and water to exit as the core tube is pushed into the sediment and providing suction to hold the sediment in place as the tube is pulled upwards. Hard objects such as rocks, oysters, or shell-hash may impede penetration to target depth. Using a larger diameter tube may help avoid the interference of hard objects. Multiple attempts needed to obtain sufficient sediment quantities for analysis should be made, moving up-current from previous attempts, until sufficient volume is collected or it is determined that piston-core sampling is not feasible in the area. Four core samples should be collected if using a 4"-diameter core tube; a total of 8 core samples will be needed if using a 3"-diameter core tube. Core samples should be collected with 18" or 24" core tubes and should be pushed into the sediment 12" to 18" into the bottom, or to the point of resistance (when the core cannot be pushed any further). A hammer (e.g., mallet or slide hammer) can be used to drive the core further into sediment, however, care should be taken not to drive the core in to the point that it cannot be pulled out by hand.

It may be necessary to conduct diver-operated coring in these situations as sample location can be chosen to avoid hard objects.

The procedure for collecting a sample using the piston corer follows:

1. Upon arrival at the station, the field team should begin by confirming the water depth with a weighted tape measure and record the depth along with station coordinates. The type of sediment felt when measuring depth should be noted.
2. It is important to collect a core sample from a stable platform. In instances of strong wind or current the boat should be anchored to maintain location and for ease of sampling. The core tube must remain as close to vertical as possible to collect an acceptable sample.
3. Decontaminate core tubes and caps using a three-step field decontamination process. Ensure that all surfaces are free of visible sediment. Thoroughly wash all surfaces with soapy water and a bristle brush and then rinse with deionized water. Follow the DI water rinse, a rinse on all surfaces with site water may be conducted. If site waters are not considered suitable for a site water rinse, deionized water may be used solely.

   Alternatively, core tubes may be pre-cleaned by the analytical laboratory and wrapped in aluminum foil. In such cases individual core tubes should be used to collect each sediment sample at a given station.
4. The piston core head is a cylindrical piece of aluminum, which has a threaded hole at one end to receive the extension poles. The first extension pole should remain attached to the piston head. Additional poles may be added to accommodate the water depth at the station. It is recommended to mark the extension rods with one-foot increments.
5. Attach the core tube to the head using hose clamps and add extension rods as needed to the other end of the head. Lower the piston core sampler into the water allowing the core

tube to fill with water. Air and water should come out of the one-way valve on the side of the piston head.

6. Gently lower the core tube until it contacts the sediment surface, being careful not to disturb the flocculent layer and keeping the tube and rods vertical. Note the location of the waters surface on the side of the extension rod and note the mark 12" or 18" (depending on the desired length of core) above the water surface. Push the sampler straight down using hand force if possible or, if necessary, a small sledge or slide hammer, until the second mark is reached.
7. Gently pull the sampler straight up removing extension rods as needed. Place a cap on the bottom of the tube when it reaches the surface of the water. The tube can then be set on the boat and the hose clamps and head can be removed. In softer sediments, the cap will need to be placed over the bottom quickly to avoid the loss of sediment from the bottom. Some loss is acceptable if the top 5 cm (2") are primarily undisturbed.
8. Rinse the outside of the core tube with water, then measure and photograph the core.
9. If the sample is considered acceptable using the criteria listed above, remove the piston core head by releasing the hose clamps. Remove the overlying water using a siphon or other similar means, taking care not to remove the flocculent layer.
10. Using a clean stainless steel spoon, collect the top 2 cm of the sample. Depending on the length of the tube and the sediment core it may not be possible to reach the sediment surface with a spoon. If the sediment is out of reach, the core tube can be cut with a hack saw one inch above the sediment surface, or the sediment can be pushed up from the bottom using an extruder until the surface sediment is reachable with a spoon.
11. Repeat the previous procedure for the collection of the next deeper sediment layer (2-4 cm) for archive.
12. Process samples in a manner consistent with the instructions in Section 4.
13. If the sample is not acceptable, remove the tube from the head, rinse clean of sediment away from sampling site and redeploy.

3.6 Sediment Collection with Diver Operated Core

1. Upon arrival at the station, the field team should begin by confirming the water depth with a weighted tape measure and record the depth along with station coordinates. When divers are sampling from a boat, anchoring is highly recommended to maintain a stable station location and provide a safe working area for divers.
2. Four inch diameter core tubes are either pre-cleaned and wrapped in foil or cleaned in the field. Cores cleaned in the field should follow a three-step decontamination procedure. Thoroughly wash all surfaces with soapy water and a bristle brush and then rinse three times with site water. Follow the site water rinse with a rinse on all surfaces with deionized water. If site waters are not considered suitable for a site water rinse, deionized water may be substituted.

3. Divers should approach from down-current of the station taking care not to disturb surface sediments. For sites with fine sediments and flocculent layers, great care should be taken to approach the site slowly and using buoyancy to settle to the bottom without disturbing surface materials. Limited visibility may require marking a station with a buoy and divers descend the buoy line to the station.

4. Once at the bottom, the diver will locate appropriate substrate for sampling, avoiding areas that are not representative of the station, areas of substantial rock, or areas that exhibit thick, dense shell hash. If water is turbid from the divers' approach, the sampler should wait until the suspended sediments near the sediment surface have settled and the water returned to its ambient condition.

5. Divers descend with four tubes and eight caps so that sufficient material can be collected in one dive. All tubes are inserted into the sediment to a depth of six inches. If needed, the core tube may be gently rotated or moved from side-to-side to facilitate insertion. Movement should not suspend the surface sediment. A cap is placed on the top of the core immediately after insertion to prevent disturbance and provide backpressure needed to retain the sediment during retrieval. The diver can then either slide his/her hand under the bottom to pull the tube up and cap the bottom or dig a small hole next to the tube to cap the tube before retrieval.

6. Cores are brought to the surface maintaining the vertical position in which they were collected.

7. Rinse the outside of the tubes with water, then measure and photograph the cores.

8. If the sample is considered acceptable using the criteria listed above, remove the overlying water using a siphon or other similar means, taking care not to remove the flocculent layer. Using a clean stainless steel spoon, collect the top 2 cm of sample. Depending on the length of the tube and the sediment core it may not be possible to reach the sediment surface with a spoon. If the sediment is out of reach, the core tube can be cut with a hack saw one inch above the sediment surface, or the sediment can be pushed up from the bottom until the surface sediment is reachable with a spoon.

9. Repeat the previous procedure for the collection of the next deeper sediment layer (2-4 cm) for archive.

10. Process samples in a manner consistent with the instructions in Section 4.

11. If the sample is not acceptable, remove caps, and rinse clean of sediment away from sampling site and redeploy.

Replicate Sample Locations

Replicate samples should not be collected in the same sampling location. The sampler operator should collect each replicate from opposite sides of the boat moving in a stepwise fashion up the sides of the boat in an up-current direction. Each sampling station should be at least 3 feet

apart. In the event that more replicates are required after the operator reaches the most up-current location on the boat, the boat should be moved at least 30 ft in an up-current direction and the process repeated.

4. SAMPLE PROCESSING.

The samplers should be maintained in an upright position until the surface sediments have been removed. Care should be taken to minimize disturbance of surface sediments.

Dewatering

On the boat, the sample should be allowed to settle for at least 1 minute. It is recommended that the dewatering occur before moving to another station as the motion of the boat can disturb the sediment. The overlying water should be siphoned off and returned to the sampling area. A concerted effort should be made to avoid the removal of flocculent material.

Surface Sample

A decontaminated spoon should be used to scoop the sediment layer out of the sampler and into a stainless steel mixing bowl.

Subsurface Sediment

Once the surface sediment is removed from a core sampler, the remaining sediment will generally be returned to the water. If additional samples are to be collected at the same station, care should be taken to release unused sediment after all samples are collected or release them into the water down current of the sample collection area. If subsurface sediments will be retained for analysis, use a hack saw to cut the core within the top inch of the remaining sediment. Alternatively, a core extruder can be used to facilitate sample collection. The core should be capped and maintained in an upright orientation until frozen for shipment and storage.

Resource Area Composites

The field team will be given the coordinates for up to eight sediment sampling locations for each study grid. The field team will visit the sample locations in the order specified, with the goal of collecting four individual samples per cell that may then be combined into two composite samples by the analyzing laboratory. The field team will collect approximately equal volumes of sediment at each location, which will be transferred into sample containers and sent to the lab under chain-of-custody.

Sample Homogenization

If homogenization is required, each sample should be thoroughly mixed for minimum of one (1) minute and should be homogenous in appearance. In particular, no separated water should be

present and there should not be significant streaking. Water may separate, particularly in sand samples. Samples should be remixed if necessary.

Sample Splits

The sediment sample should be divided into three sample containers (grain size, deep, and surface samples) with a capacity of 250 mL. These containers should be filled to a minimum of 50%, and preferably 75% capacity to assure enough sample container capacity during frozen storage.

Decontamination

Wash the spoon and stainless steel mixing bowl with dilute Alconox between samples. Thoroughly rinse all surfaces with deionized water.

Labels

The sample container lids should be labeled immediately before filling with the sample location, date and time. An identical record should appear in the field log with any additional information that is required under the QAPP. The samples will be transferred with a complete chain of custody to the sample intake team as soon as possible.

Sediment analyses will be performed in accordance with the NRDA Analytic Chemistry AQAP as summarized in Table A-3.

Table A-3. Sample Analyses and Sizes.

		Sample Size (g wet)		Collected	
Test	Reference Method	Min	Ideal	(mL)	Sample Storage
Grain Size	PSEP protocol	100	175	187.5	Refrigerated
TPH/THC	EPA 8015				
PAH	modified EPA 8270d	50	75	187.5	Frozen
Biomarkers	modified EPA 8270d				
TOC	EPA 9060	0.5	2		
Total		150.5	252	375	

5. QUALITY CONTROL

The quality control procedures for this method include three primary parts: staff training, equipment blanks, and field duplicates.

Staff Training

The field sampling staff shall read this SOP and be shown how to properly operate all field sampling equipment by an experienced staff member before performing these tasks without supervision. This training will include specific guidelines for how to accurately, precisely, and safely load and trigger the sampler without placing any appendages in the mouth of the sampler.

Equipment Blanks

Whenever possible, the field team should use disposable sampling equipment (e.g. polycarbonate core liners and gloves) during the collection of each sample. It is recommended that one equipment blank be collected for each 20 samples. The equipment blank consists of distilled or deionized water pored over reused sampling equipment (e.g., stainless steel bowls and van Veen grab samplers). The rinsate shall be collected in a 1 L bottle. This water blank should be analyzed by the same hydrocarbon testing methods requested for the associated sediment samples

Field Duplicates

It is recommended that each field team collect one field duplicate for each 20 samples. The field duplicate should be collected in a manner identical to the original sediment sample (e.g., a composite of two sampling locations within an oyster bed). The field duplicate should be analyzed by the same hydrocarbon testing methods requested for the parent sediment samples.

D. SOP for Decontamination Procedures for Sampling Equipment
Adapted from "the Standard Operating Procedure Decontamination Procedures for Sampling Equipment MC252 Fish Technical Work Group Plans," August 24, 2011

1. **Scope and Applicability**

This Standard Operating Procedure (SOP) describes equipment and field procedures necessary to properly decontaminate equipment utilized for the MC252 2011 Oyster Quadrat and Sediment Sampling Plan under which sediment and tissue sampling are conducted. This process is designed to minimize the potential for constituent migration and/or cross contamination. This procedure does not apply to personnel decontamination.

2. **Summary of Method**

The objective of these multimedia sampling programs is to determine and quantify the presence of oil-related chemicals in oyster habitat and as well as in oyster tissues. Decontamination procedures appropriate to the oil-related chemicals being assessed may improve the prevention of cross contamination. This SOP presents an adaptive approach to decontamination that ensures sufficiency of decontamination while minimizing the use of and personnel exposure to solvents.

3. **Equipment and Supplies**

- PPE (including disposable Neoprene gloves, chemical splash goggles; see Section 4.0 below for additional information including safe work practices)
- Small dry chemical Fire Extinguisher (BC or ABC Rated - 5 lb or larger)
- Bristled Brushes compatible with the solutions being used
- Low Phosphate Detergent (Alconox or Liquinox), diluted in accordance with instructions provided with the product.
- Acetone and Hexane (pesticide grade or better stored in ETFE Bottles)
- Distilled/DI water
- Designated solvent-compatible container for collection of decon waste/rinsates (HDPE ok for hexane and acetone, if acetone concentrations are less than 5%. Otherwise a PTFE liner is needed).
- Secondary containment vessel such as a cooler that can be closed to reduce the likelihood of spills and reduce volatilization
- Clean Ambient/Tap water source
- Wash/rinse tubs compatible with the solutions being used
- Specified area of vessel for decon away from other contaminant sources and other personnel
- If collecting a rinsate blank, small container appropriate for the collection
- Field documentation materials

4. **Health and Safety**

Health and safety hazards associated with this procedure can be mitigated by the following engineering, administrative, and PPE controls:

HAZARD	CONTROL(S)
Bodily injury due to pinch points or dropped equipment	• Leather gloves and steel-toe boots should be worn while equipment is being handled • Equipment safety features (e.g., lock pins) should be engaged while equipment is being handled
Vapor inhalation	• Use solvents only in well-ventilated areas • Remain upwind of solvent decon work • Advise other workers in the area of the nature of your task and ask them to remain upwind
Skin irritation	• Don proper chemical-resistant gloves (disposable Neoprene 5ml or greater thickness) prior to handling organic solvent • Rinse solvent from gloves before removing • Promptly wash any areas of skin which may have encountered contact with organic solvent and always wash after completing work with hazardous materials
Eye contact	• Don chemical splash goggles prior to retrieving and handling organic solvent • Do not use solvent wash bottle near face
Fire	• Store organic solvents in approved, leak-proof containers (ETFE plastic for either solvent) in a cool, shaded area; do not store in direct sun • Do not smoke near solvent storage or work areas • Do not use or place solvent near flame or other heat source • 5- or 10-pound dry chemical fire extinguisher (Type BC or Type ABC) should be readily accessible during the decon process
Solvent spill	• Place equipment to be decontaminated in containers to capture rinsate • Inspect solvent containers for leaks prior to handling • Have an organic solvent spill kit available and near solvent storage and work areas; workers should be trained in spill kit use

	before the start of each mission
Environmental detriment	• Keep solvent bottles tightly capped to prevent leakage and minimize vaporization. Store in secondary containment vessels • Promptly clean spilled solvent with paper towels and discard in solid used waste container • Maintain solid used materials (e.g., paper towels, disposable gloves, etc.) in a bucket or other container to prevent litter • Promptly replace lids onto rinsate buckets and secondary

NOTE: The above information was determined from job hazard analysis of the work tasks

5. Decontamination Procedures

Levels of Decontamination Procedures and their Selection
All equipment and non-disposable materials that directly contact a sample medium shall be must undergo Level 1 Decontamination (see below) or be pre-cleaned by the manufacturer, in compliance with the protocols described here.

The Level 1 Decontamination procedure shall be the default decontamination procedure for all nondisposable equipment, followed by Level 2 Decontamination when applicable. The observation of oil in the general vicinity of the sampling does not necessitate Level 2 or Level 3 Decontamination (use of solvents; see below), but Level 2 or Level 3 Decontamination can be used at the field crew's discretion.

Level 3 Decontamination must be used when Level 1 and Level 2 Decontamination procedures are not successful (i.e. visible oil is still observed on the equipment or the equipment rinsate).

Level 1- Default decontamination procedure

Scrub[6] all equipment and parts with a dilute detergent mixture and rinse with deionized or distilled water. Inspect the equipment and rinse water for signs of residual oil, other contaminants, or incomplete decontamination.

[6] The full decontamination process using detergent washing procedures is described below.

Level 2 – Inspection and secondary decontamination

Whenever, after the Level 1 Decontamination procedure, there remains some evidence of incomplete decontamination and residual oil (i.e. sheen in rinse water, dark spots on net, etc.) the field team shall repeat Level 1 decontamination.

After the Level 1 Decontamination procedure is repeated, the equipment and rinse shall again be inspected. If after visual inspection there remains evidence of incomplete decontamination and residual oil (i.e. sheen in rinse water, dark spots on the net, etc.) the team shall utilize small quantities of solvents to spot clean the area of residual oil. The decontamination procedures using two solvents are described below.

Level 3 – Expanded solvent decontamination procedures

If after the Level 2 Decontamination procedures the field team determines the decontamination procedures were not adequate, the field team shall cease using the sampling equipment. The equipment shall be isolated and secured while on the work boats. More thorough decontamination, including additional detergent washing, additional solvent spot treatment and/ or expanded solvent treatment, can be conducted upon returning to shore.

Specific Protocols

These protocols are to be followed for all sampling apparatus (e.g., sediment collectors, nets, mixing bowls, etc.). Sediment samples collected for grain size analysis shall require the default procedure only.

All sampling devices between sample collections

- Collect the samples following the Work Plan's sampling protocol
- Wash and scrub with a clean mixture of distilled/DI water and low phosphate detergent
- Rinse equipment with distilled/DI water
- Inspect devices and rinse water; if sheen or oil is observed, repeat the above steps; if not, decontamination is complete
- If sheen or oil is observed after a second decontamination with water and detergent, proceed to the solvent rinsing steps below

Oil/sheen observed after repeated decontamination with water and detergent scrub

- Wash and scrub with a clean mixture of distilled/DI water and low phosphate detergent
- Rinse equipment with distilled/DI water

- Use a ETFE bottle to apply Acetone sparingly[7] to the piece of equipment being decontaminated
- Use a ETFE squirt bottle to apply Hexane sparingly to the piece of equipment being decontaminated
- Thoroughly rinse with DI/distilled water

Oiled rinsate must be collected in the designated solvent-compatible container. Keep a lid on this container at all times while not in use and apply the designated rinsate label to the side of the container. These materials will be turned over to appropriate personnel for disposal. Collect non-oiled rinsate in a bucket with lid for disposal.

NOTE: In the event that the total duration of solvent application for either Acetone or Hexane reaches 24 minutes cumulatively over the course of your work day, please discontinue solvent use for that day and contact your Supervisor and Project Safety for further direction.

6. Storage and Disposal of Chemicals and Chemical Waste

Solvents and rinsates will be handled following the specific guidelines listed below:

Solvents

- All solvents will be transported in small amounts (500ml-1000ml)
- All solvents will be transported/stored within closeable secondary containment to prevent spills and volatilization
- Keep all solvents and secondary containment as cool as possible
- Do not store solvents or rinsate materials in vehicles or hotel rooms, utilize the storage facility identified for your sampling crew – acquire the necessary solvent amount in PTFE/Teflon squirt bottles from the storage facility each morning, and return the unused portion and containers after the sampling day

Rinsates Containing Oil and/or Solvents

- Collect all rinsates in the designated solvent-compatible container with the appropriate label on the side
- Place rinsate containers in a secondary containment system to reduce the likelihood of spills and prevent volatilization
- All rinsates containing oil and/or solvents will be transported by authorized persons to the appropriate waste disposal site
- All rinsates will be captured in the same container.[8]

[7] Use the minimum possible to remove the contamination. Teams are limited to 1000ml of each solvent and 24 minutes of total use per day. Delivery rate is also limited by the nozzle size of the squeeze bottles.

[8] Diluting the rinsate from level 2 with the rinsate from level 1 is a key safety factor, reducing both concentration and volatility.

Rinsates Containing Water and/or Low-phosphate Detergents

- Rinsates containing only low phosphate detergents and water will also be containerized and transported to the appropriate waste disposal site
- Place rinsate containers in secondary containment during transportation and storage to reduce the likelihood of spills

Appendix B: OYSTER SAMPLE ID NAMING CONVENTION

NOAA NRDA Sample Format:

- **LocationCode – DateCode - Matrix Leader# Sample#**
    - 6-digit Location code (from maps located on www.noaanrda.org. These should be the NRDA Grid location code rather than the SCAT location code);
    - 5-digit date: year letter and mmdd (A=2010, B=2011);
    - Matrix letter (T = Tissue or S = Sediment);
    - 2 or 3-digit leader code; and
    - 2-digit sample number.
- EXAMPLE: **LAAM24-B0502-TA102**
    - LocationCode = LAAM24;
    - Date = 5/2/2011;
    - Matrix=Tissue;
    - Leader code = A1;
    - Sample # = 02.

Additional Information for Oysters:

Field Teams

- We will be numbering each sample sequentially *within each GCID and by sample type* (e.g., each GCID will have four quadrats numbered 01Q through 04Q, and any dredge samples should be numbered starting with 01DR.) This information will go in the "Sample #" section at the end of the NOAA NRDA required tag.
- Grid Cell ID – the Grid Cell ID number (e.g., 0024, 3989) will be added to the sample ID immediately preceding the sample number so that the site can be identified. The Grid Cell number is not unique across states, but with the state abbreviation embedded in the location code the value is unique. *Use leading zeros to ensure that the GCID is always four digits.*

- Tissue Subtype – In addition, because there are several different tissue sample types, we will add an identifier after the sample number that will indicate the sample type for tissue samples.
    - Q = quadrat sample;
    - L = larval sample (add LS for surface, LM for mid water column or LB for bottom); and
    - SP = settlement plate.
    - Example: **LAAM24-B0502-TA10024-01Q**
- Sediment Sampling Method – Add an identifier after the sample number to indicate the sampling method used and the category of sample
    - C = core sample
    - G = grab sample
    - Example: **LAAM24-B0502-SA10024-01C**
    - GS = grain size sample
    - S = shallow sediment sample
    - D = deep sediment sample
    - Example : **LAAM24-B0502-SA10024-01CGS or LAAM24-B0502-SA10024-02CS**
- All additional information describing the samples will be recorded in the "Sample Notes" field of the NOAA NRDA sample collection forms (see OysterExamples.xls). This additional information differs by sample type.
    - Quadrat oysters
        - Site Name
        - Quadrat Number
    - Sediment
        - Site Name

Lab Teams

- **Quadrat subsamples**

- Contaminant subsample
    - Keep original sample name for the quadrat from the site and add "-CT", e.g., **LAAM24-B0502-TA10024-01Q-CT**

- The labs will track the sample ID changes, splits and composites in a sample bridge template and upload to noaanrda.org site, under instruction from the data management TWG. In addition, the labs will upload result information to the www.noaanrda.org site on a frequency agreed upon by the lab and the data management TWG.